# Care and Conservation of Natural History Collections

Title: Insects and other invertebrates

Author(s): Walker, A. K., Fitton, M. G., Vane-Wright, R. I. & Carter, D. J.

Source: Walker, A. K., Fitton, M. G., Vane-Wright, R. I. & Carter, D. J. (1999). Insects and other invertebrates. In: Carter, D. & Walker, A. (eds). (1999). *Chapter 2: Care and Conservation of Natural History Collections.* Oxford: Butterwoth Heinemann, pp. 37 - 60.

URL: http://www.natsca.org/care-and-conservation

# 2

# Insects and other invertebrates

**Annette K. Walker*, M. G. Fitton, R. I. Vane-Wright and D. J. Carter**
*Entomology Department, The Natural History Museum, Cromwell Road, London SW7 SBD, UK*

**Formerly of the International Institute of Entomology.*

## Introduction

Invertebrates, particularly insects and shells (molluscs), have been popular with collectors since the seventeenth century and there is an extensive historical record of preservation methods and display. Among the oldest insect collections surviving are those of Leonard Plukenet and James Petiver. These two collections formed part of Sir Hans Sloane's collection which in turn became the foundation of the British Museum's natural history collections, now The Natural History Museum in London. Leonard Plukenet was an eminent seventeenth century botanist so it is hardly surprising that the specimens he collected around 1696 were pressed like flowers and attached to sheets of paper hound as a book (Plate 6). Many of these specimens are still in remarkably good condition, with colours only slightly faded. James Petiver (1663–1718) was an apothecary who made an important early collection of insects. However, the collection was in a poorly curated state when Sloane purchased it and he employed a curator, Cromwell Mortimer, who mounted many of Petiver's specimens individually, sandwiched between thin sheets of mica (Plate 7). Other parts of Sloane collection were housed in shallow, glass-lidded cabinet drawers which resemble those used by entomologists today (Fig. 2.1). The main lessons to be learned from these old collections are that specimens stored dry, away from light and free of pest attack, will survive for at least 300 years with minimal deterioration.

The possibilities of preserving insects dry were obviously appreciated by Plukenet and Petiver, as was the need also to protect the fragile specimens from damage. By the time Linnaeus published the tenth edition of his

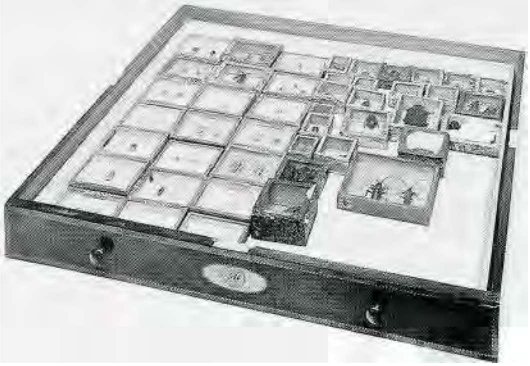


**Figure 2.1** The Sloane Collection. This part of the collection illustrates a further advance over specimens being pressed flat. Here they are housed in small, shallow, glass-topped containers. The specimens are secured in place with adhesive; no pins are used. The specimen containers themselves are kept in shallow, glass-lidded cabinet drawers, which resemble those used today.

*Systema Naturae,* some fifty years later, the techniques of pinning insects and arranging them in cork or wax-lined, glass-lidded drawers had become established. At first data labels were pinned into the drawers separately from the specimens and few species were represented by more than one or two individuals. It was not until towards the end of the eighteenth century that the need to examine and preserve series of specimens and to attach a label to each began to be appreciated. This system has withstood the test of time and is still the basis of present-day methods.

The study of insects, arachnids and other invertebrates poses special problems because they occur in vast numbers, both as species and individuals. They are often collected in large numbers using traps, nets and other means of sampling. Perhaps as many as 30 million distinct species exist today and most show at least some seasonal, geographical, polymorphic or other form of variation; and, in addition, each individual passes through a series of distinct developmental stages during its life. The result is that even a modest collection comprises thousands of individual specimens. The average scientific value of an individual specimen may be relatively low, but is vastly increased if it is part of a well curated and organized collection. However, individual specimens, for example primary types, may be extremely important or have historical value and some rarities may have high financial value as collectors' items.

Each specimen has to be examined and identified in the light of existing knowledge (and often conflicting opinion), and then re-examined and re-interpreted as new understanding of the natural classification is gained. Unfortunately, the difficulties associated with the identification of invertebrates are acute and there are other problems associated with the need to deal with large numbers of specimens: a minor difficulty at the individual level can create a major curatorial headache by simple multiplication. All this means that the development and curation of a comprehensive collection of even a single group, such as butterflies or mosquitoes, or just a limited geographical area for a regional museum, is an enormous task.

The different requirements and techniques involved in the care of collections of invertebrates involve all major aspects of collections management, and these are covered in this chapter. The first part deals with arthropods, particularly insects and spiders, but will also contain information of value to those working with other invertebrate collections. More specific recommendations will be found in the subsequent section on other invertebrates and in Chapter 5 on fluid preservation.

As far as possible, the arrangement follows the sequence of events involved in the formation of a collection but collecting and sampling methods have been omitted as they have little bearing on a specimen's subsequent care. The chapter pays particular attention to the problems faced by those who are dealing with existing collections.

Information on the preparation and curation of insects and arachnids can be found in many existing texts. Recent general treatments which we recommend are Walker and Crosby (1988), Martin (1977) and Upton (1991). These works include detailed information for each taxonomic group and developmental stage. Gilbert and Hamilton (1990) is a thorough guide to information sources in entomology. Other invertebrates are dealt with by Lincoln and Sheals (1979) and Wagstaffe and Fidler (1955).

## Arthropods

Although the dry preservation of many arthropods is permitted by their tough, rigid external skeleton', it also restricts the ways in which they can be studied. Moreover, the soft nature of the cuticle of many groups, or the need to macerate specimens (dissolve and clear soft tissue) to reveal fully hard-part anatomy, demands other means of preservation. As a consequence three major (and often complementary) modes of preservation are widely applied: dry (usually on pins in 'air-tight' containers), in liquid (usually in vials of alcohol), and as microscope preparations on glass slides.

### Specimen preparation, handling and labelling

This section comprises a brief account of techniques involved in specimen preparation

based on recommended, current practices. Details of methods only applicable to material already in collections are given below in the section on Remedial Conservation (pp. 54).

***Pinned material***

Traditionally, insects (especially adults) are preserved dry, impaled on pins. Dry specimens are fragile and easily damaged if handled directly and pins have proved to be a simple, effective means of providing for safe handling, storage and attachment of labels. This method has also been employed in the past for some crustaceans but is no longer used as specimens easily become disarticulated and are difficult to reassociate. However, many such old collections still exist and these require careful handling.

*Relaxing*

Freshly killed specimens which are still relaxed can be mounted without any special treatment. To avoid damage, those which have dried out and stiffened or have been stored dry between layers of tissue or entangled in cotton wool, need to be relaxed before they can be mounted. This is a very simple process carried out in a humid chamber, commonly called a 'relaxing box', which can be any suitable air-tight plastic or glass container (early relaxing boxes were made of zinc). The technique is as follows:

Cover the bottom of the box with a thick layer of cotton wool or cellulose wadding which is almost saturated with water.

2. Add a few drops of phenol or a few crystals of chlorocresol or thymol (or if these are not available a little domestic disinfectant) to stop mould growing but never place the specimens directly on this wet layer because they will be damaged by excess moisture.
3. Gently lay the specimens on filter-paper in a shallow dish, such as a Petri dish or plastic lid.
4. Leave overnight or even up to a few days if the specimens are large and robust.
5. Check the specimens frequently as they will deteriorate quickly if left in the humid atmosphere for too long.
6. Do not leave the relaxing box in the sun or near a source of heat as condensation will form on the underside of the lid and drop on to the specimens.

Ensure that labels in the relaxing box are not written in water-soluble ink. Specimens that are required for molecular studies should not be relaxed in this manner as it causes degradation of DNA (Carter *et al.*, 1997). In such cases, sample material should be removed before relaxing.

Specimens that have been stored dry on layers of tissue are best relaxed on the tissue. Gently peel off the top layer of tissue in the storage box and carefully, with a brush, remove any specimens that may have adhered to it. Then place the entire layer in the relaxing box on a flat platform (a piece of perforated zinc or `expanded' stainless steel bent down at each end to prevent the tissue from coming into contact with the damp surface).

*Handling*

Watchmakers forceps with very fine points are the best type for direct handling but care must be taken because the points are hard and inflexible and can very easily break fragile parts. With practice the pressure applied to a specimen can be controlled easily by cushioning the jaws of the forceps by placing the edge of a finger between them (Fig. 2.2). The points of watchmakers forceps easily bend or break off but damaged tips can be reground with a fine sharpening stone (used under a

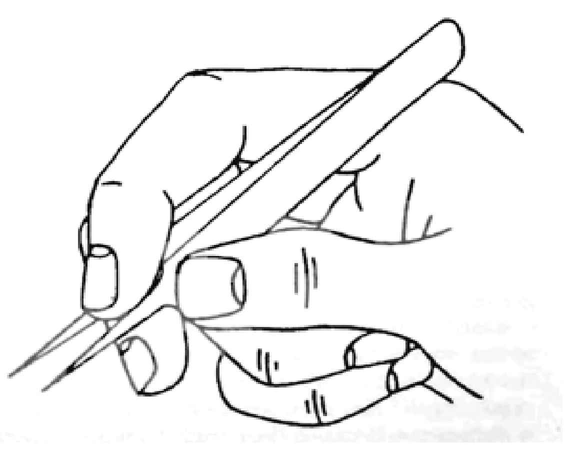


**Figure 2.2** Cushioning the pressure applied to a specimen when using watchmakers forceps.



**Figure 2.3** Fine-tipped, slender forceps for handling delicate, unmounted, dry specimens.



**Figure 2.4** Blunt-nosed forceps for handling larvae.

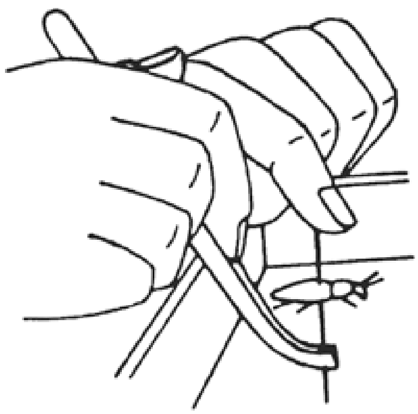


**Figure 2.5** The use of entomological pinning forceps to grip the shank of very fine pins and steady the specimen at the same time.

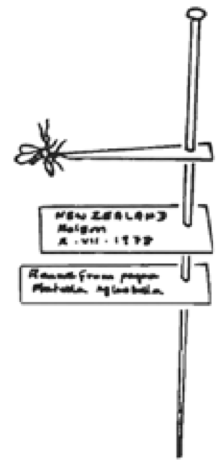


**Figure 2.6** A small insect glued to a card point

dissecting microscope to ensure that the tips are ground to an equal length and close properly). Two types of flexible, lightweight forceps are also useful for handling specimens directly: fine-tipped, slender forceps (Fig. 2.3) are ideal for delicate, unmounted, dry specimens and blunt-nosed forceps for larvae (Fig. 2.4). Philatelists forceps can be used for handling insects stored dry in paper triangles. Fine, soft paintbrushes are useful for handling unmounted specimens whether dry, relaxed or in ethanol. Sable brushes are best, but less expensive camel-hair brushes are almost as good.

Entomological 'pinning' forceps are curved at the end and designed to grip the shank of the handling pin of a pinned insect at a convenient angle (Fig. 2.5). Never grip the pin head with forceps because they may damage it. Also forceps tend to grip some pin heads at an oblique angle, making the pin and specimen spin off violently, often causing irreparable damage. Although pinning forceps are essential equipment, the handling pins of some pinned specimens can be held safely between finger and thumb.

*Mounting*

Large, robust insects such as beetles, moths, large flies and wasps are usually mounted by being directly impaled by the handling pin. Smaller insects are dealt with in a variety of ways. They may be glued to a card point (Fig. 2.6) or rectangle (Fig. 2.7) which is supported by the handling pin. Small Diptera and microlepidoptera are usually pinned with a very fine short stainless steel pin ('minuten' or `micro-pin') which is then held in a stage supported by the handling pin (Fig. 2.8). Another technique, popular for medium sized Hymenoptera and Diptera, is to glue the insect directly to the shaft of the handling pin (Fig.

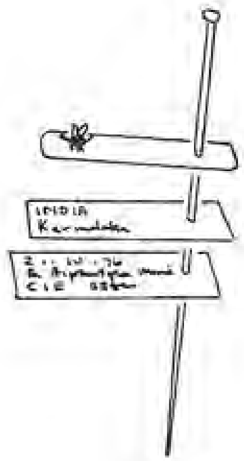


**Figure 2.7** A small insect glued to a card rectangle

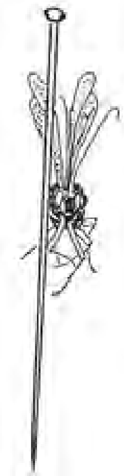


**Figure 2.9** A medium sized Hymenoptera specimen glued directly to the shank of a handling pin.

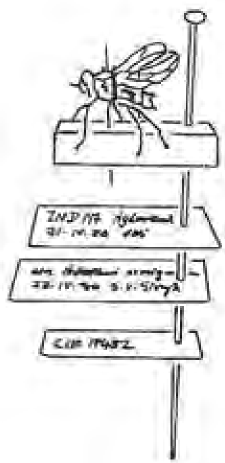


**Figure 2.8** A small dipterous insect pinned with a 'minuten' or 'micro-pin' held in a stage support.

2.9). Modern insect pins are a standard length (38 mm, known as `Continental' length in Britain) and made from stainless steel. They have a spherical head, often of nylon, and come in various thicknesses. Thinner pins such as '0' and `1' are used for smaller, more delicate specimens and the thickest ('5') for very large insects, cards and double mounts. The finest pins ('00' and finer) are impractical in use and are easily damaged.

Some cheap `stainless steel' pins are of very low quality (poor points and inferior metal) and may be susceptible to corrosion. If possible, *English* entomological pins should also be avoided. They come in a variety of shorter lengths which do not leave adequate space for safe handling or labels and they are not always made from stainless steel. All cheaper steel, lacquered or blackened or nickel plated brass pins are subject to problems of corrosion and verdigris. Unfortunately cabinet drawers have often been designed to take short, English-length pins and they are too shallow for standard 38 mm pins. If such drawers need to be used then headless, stainless steel pins (of the maximum possible length) are probably the best, easily available compromise. Some drawers will accommodate long pins at a slight angle, and this is preferable to cropping the pins themselves.

Water-soluble vegetable glues are recommended for attaching insects to card points and rectangles. Recipes, based on natural gums such as gum arabic and tragacanth, can be found in various collecting manuals. Tradition, personal preference and availability all play a part in the choice, but the possibilities of the specimen later needing to be remounted or removed for more detailed study

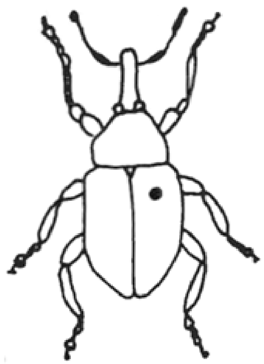


**Figure 2.10** Pinning position for Coleoptera.

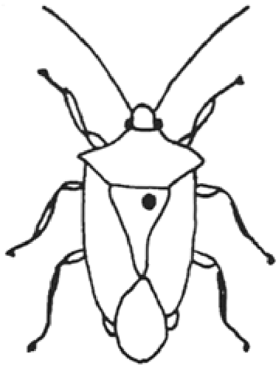


**Figure** 2.11 Pinning position for Heteroptera.

should be kept in mind (see, for example. Noyes, 1982). A commercially available fishbased glue Seccotine is used by many entomologists. Such organic adhesives must be used with caution for specimens intended for molecular studies as they may cause contamination. Shellac (an insect-based resin) is usually used to glue insects direct to the shaft of the pin (in the past some workers have used clear nail varnish for this purpose but there is some question over its archival nature – care should he taken to use 'conservation grade' adhesives that will not acidify or otherwise deteriorate in a way that may damage the specimen – reversibility is essential). Card for mounts should be good quality three– or four–ply Bristol board. Stages for double mounting of micro-pins were traditionally made of strips cut from dried Polyporus fungus or elder pith but a good modern substitute can be made from Plastazote (cross-linked polyethylene foam) provided that it is sufficiently dense to be rigid. This synthetic substitute has the advantages that it is consistent in texture and does not dry out and become brittle.

Mounting methods must be cost-effective, economical in terms of initial preparation time, and take into account longer-term factors such as the use specimens will be put to, the space taken up in storage containers and risk of damage in handling. Care must also be taken to ensure that the methods used do not damage or obscure important features. For direct pinning, there is an accepted pin position for each group, but as a general rule it is through the mesothorax slightly to the right of the midline to avoid important features on the dorsal and ventral surface. Coleoptera are pinned through the right elytron (Fig. 2.10) and Heteroptera through the right of the scutellum (Fig. 2.11). In Lepidoptera the pin goes through the middle of the thorax (Fig. 2.12). There are many variations on the basic techniques and texts on collecting and on individual groups should be consulted for detailed information. No two entomologists agree totally on methods, though some methods, such as spreading the wings of butterflies and moths (Fig. 2.13), are almost universally accepted. General accounts covering all orders include Walker and Crosby (1988), Martin (1977) and Upton (1991). Specialists often develop their own techniques, applicable to only a small group or requiring more time and skill than can be justified by the average collector or curator. For example, the technique described by Noyes (1982) for mounting small chalcid wasps is unlikely to produce satisfactory results without a lot of practice.

Most insects, even quite large specimens, can be air-dried without problems. However, specimens may need to be warmed to accelerate the drying process and for some groups, for example Orthoptera. Specialists recommend that large specimens be stuffed, but care is needed to avoid damaging important characteristics. Marshall and Haes (1988) give an account of this technique.

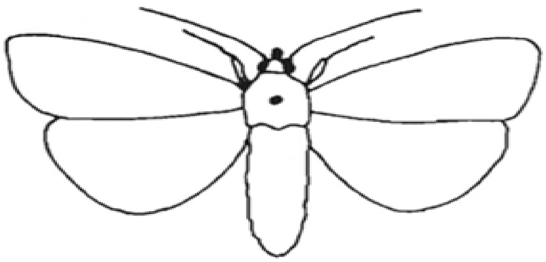


**Figure 2.12** Pinning position for Lepidoptera.

*Dry mounting specimens from alcohol*
Many winged insects, such as Hymenoptera, are routinely stored in ethanol and then removed for dry mounting. The technique is relatively straightforward and is described in Gauld and Bolton (1988). The important points include: rinsing specimens in clean ethanol before allowing them to dry out, making sure that wings are not crumpled, and mounting them before they start to become brittle.

Specimens of some groups which would not normally be stored in ethanol, for example adult Lepidoptera and hairy insects such as bees and some Diptera, can be `recovered' to make dry, pinned mounts. The techniques (which are best carried out in a fume cupboard) generally involve treatment in volatile organic solvents such as acetone or ethyl acetate followed by some grooming of the specimen. Instructions are given by Sabrosky (1966). Milliron's (1971) account of cleaning bumble bees is also useful.

Also very fragile specimens, such as small Hymenoptera and Diptera, which may collapse or distort when removed from alcohol can be treated either by critical-point drying (see below) or by the use of hexamethyldisalizane (HMDS). The latter involves transferring specimens from 100% alcohol through two or more changes of HMDS before allowing the specimens to air-dry in a fume cupboard (HMDS is a dangerous substance and must be used with great caution). Full details are provided by Brown (1990, 1995) and Cowan (1995).

*Freeze-drying*
Freeze-drying is a technique that is particularly useful for display purposes but is also valuable for dry preservation of soft bodied arthropods such as spiders and insect larvae for reference collections. Frozen specimens are dried below freezing point under a vacuum so that ice crystals are gradually sublimed into water vapour and removed (see also Chapter 1 on vertebrates). For groups such as dragonflies, colour retention can be quite outstanding compared to other techniques. However, specimens freeze-dried after storage in ethanol discolour, often split and do not look lifelike. A general account of freeze-drying techniques is provided by Hangay and Dingley (1985). The equipment required to carry out this process is quite costly. Larvae less than 10 mm long are difficult to freeze-dry unless special precautions are taken to keep them below freezing point throughout the procedure. Critical-point drying is often better for very small specimens (see below).

While preparing specimens for freezedrying, place the freeze-drying container in a

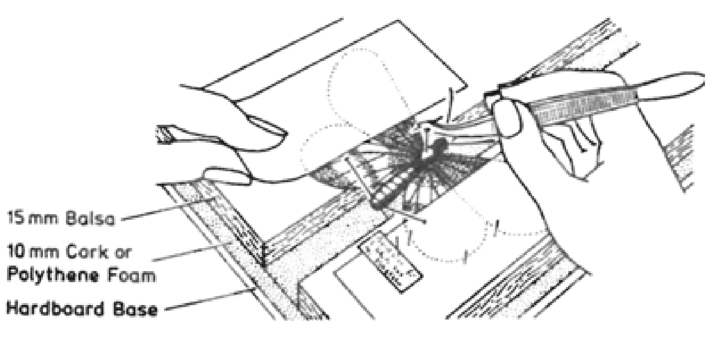



**Figure 2.13** Setting Lepidoptera (after Upton and Norris, 1980).

deep-freeze to cool it. Lower specimens into liquid nitrogen. Larvae that tend to react violently when handled with forceps can either be anaesthetized or lowered into the liquid nitrogen while they are still feeding on their host plant (which can also be freezedried). After the vigorous boiling reaction has slowed down, remove the specimens quickly into the cold freeze-drying container. Place the container into the freeze-drier as soon as possible or the specimens will start to thaw. The time required for complete drying depends on the size of the specimen and must be determined by practice. A detailed account of freeze-drying techniques for spiders is provided by Moore (1977). Freeze-dried specimens are extremely brittle and must be handled with care.

*Critical-point drying*

The technique of critical-point drying allows small specimens, such as Hymenoptera and. Hemiptera, to dry without the body collapsing or losing further colour. Appendages remain more flexible than those of air-dried specimens and are therefore less likely to be damaged, but colours are slightly dulled (Craig and Craig, 1987). This technique involves passing specimens through an intermediate fluid (acetone or 100% ethyl alcohol) and then into a transitional fluid (usually liquid CO2 or Freon), before subjecting them to critical temperature and pressure to effect drying (Gordh and Hall, 1979). It requires specialist equipment and must only be carried out by trained personnel. Critical-point dried specimens can be stored dry in vials or layered between tissue if they are not mounted immediately.

*Ultrasonic cleaning*

Some soil-inhabiting arthropods may be covered with dirt and debris and trap catches sometimes become choked with Lepidoptera scales and other debris. Such material can be cleaned using an ultrasonic cleaner. Specimens can be treated in their storage fluid (usually ethanol), but best results are obtained by placing them in a warm solution of detergent/wetting agent (e.g. Decon 90) in deionized water in a closed vial which is then dipped into water in the tank of the ultrasonic cleaner (see also Appendix IV which considers a case study). Alternatively, place specimens in a basket and immerse them directly in a detergent solution in the tank. Tests should be made to ensure that the power of the ultrasonic cleaner is not so great as to damage or destroy the specimens, and best results are achieved when all the specimens are of similar size and robustness. The time required for cleaning will depend on how large and dirty the specimens are, but the time will usually be in the range of a few seconds to two minutes. Shear and Levi (1970) give useful hints for ultrasonic cleaning of spiders and myriapods. Ultrasonic cleaners should be used with caution: see Chapter 9 on Policies and procedures, and Health and safety, p. 190).

*Dissections and preparations from pinned specimens*

Often parts of a specimen which have been removed for examination, for instance genitalia, are not made into permanent preparations on microscope slides but are glued on to the specimen mount or stored in glycerine in microvials (Deitz, 1979). These glass or polyethylene microvials usually have stoppers of cork or neoprene, respectively. The vial is held at an angle below the specimen, by the pin passing obliquely through the stopper to avoid glycerine coining into contact with the stopper. However, over time corks will inevitably come into contact with the glycerine and deteriorate and they are not recomended. Moreover, they are notoriously inconsistent in quality and break with age.

***Microscope slides***

Textbooks on microscopic techniques usually do not discuss in depth the preparation and mounting of small organisms such as small insects and mites. The exact requirements vary but the accounts by Mound and Pitkin (1972), Robinson (1976) and Noyes (1982) cover important points to consider. Some mountants and mounting techniques employed in the past have been unsatisfactory, leading to degradation of the mounts and, in some cases, destruction of the specimens (Upton, 1993). In a comprehensive survey of slide-making techniques, Brown (1997) draws attention to problems with mountants such as gum chloral, which tend to crystallize or blacken with age.

This may be due either to incorrect formulation of the mountant or variation in preparation technique. Many mountants should be considered only suitable for temporary preparations. Resin-based mountants such as Canada balsam and Euparal, which have stood the test of time, are probably the best option for permanent mounts, although it is possible that some modern synthetic mountants will prove to be of archival quality.

Glass microscope slides are quite robust but must still be handled carefully. As long as the mountant is not soft, the coverslip and slide can be carefully cleaned with lens tissue or a soft lint-free cloth (old handkerchiefs are ideal).

### *Plant and other material associated with insects*

Collections of nests, leaf mines, galls, wood borings and scale insect and whitefly `pupae' are usually stored dry. Leaves with mines can be pressed and attached to herbarium sheets (see Chapter 3 on Vascular plants). Scale insects and whitefly pupae can be stored in paper packets (archival quality). Galls, wood borings and small nests can also be stored in packets or unit trays in drawers. As large nests can provide a rich food source for insect pests, they should be dried and sealed in polythene bags and ideally stored in pestproof cupboards. Cast skins, pupal cases, cocoons and host remains of parasitoids associated with individual adults are best placed in a gelatin capsule impaled on the same pin as the specimen. If they are glued to a slip of card they sooner or later become detached and parts such as cast skins within cocoons fall out and are lost.

### *Fluid Preservation*

Although this subject is dealt with in Chapter 5 (Fluid preservation), a few details are worth mentioning here. Soft bodied arthropods such as caterpillars and spiders, which usually collapse and become useless for study if dried, are best preserved and stored in ethanol (alcohol) diluted with distilled water to 70—80% by volume. Some hard bodied arthropods such as Crustacea and some mites are also best stored in ethanol. IMS (see p. 131) is a cheaper alternative to pure ethanol but may be unsuitable for material intended for DNA study. As ethanol is a fixative as well as a preservative, most specimens can be collected directly into it. However, larvae of some insects tend to blacken if treated in this way and they should first be fixed by immersing in boiling water for one minute before drying and transference to alcohol. Specimens should be transferred to fresh alcohol after a few days to compensate for dilution by body fluids, which can lead to deterioration and ultimate decay.

Long-term storage in ethanol is not recommended for some groups. For example, aphids, mealy-bugs, thrips and some small Diptera should be slide-mounted and small Hymenoptera critical-point dried as soon as possible. Many insects in ethanol slowly deteriorate, becoming decolorizecl, increasingly brittle and in some cases even losing fine cuticular structure (such as microtrichia). It is recommended that specimens stored in ethanol for a long time should ideally be kept in a deep-freeze at -20 to -30°C.

In countries where it is difficult to obtain ethanol, alternative preservatives may be used such as isopropanol, although this does not have identical properties and may tend to make specimens brittle. For spiders, a preservative based on propylene phenoxetol has been used successfully; details are given by Hillyard (1989). Some old collections may be stored in formaldehyde (which is now a known carcinogen) and require special precautions in handling and transfer to alcohol (see Chapter 5, Fluid preservation).

### *Specimen labelling*

Specimen labels may conveniently be attached to pins and microscope slides although space on the pin or slide is limited. Often abbreviations have to be used and they should follow some accepted system. Larger, more comprehensive labels may be added to material preserved in liquid. Labels are generally of four kinds:

1. A locality label which gives basic collecting data. The accepted format presents information in increasing order of precision, for example country (in capitals), county, town, co-ordinates and altitude, followed by the date and collector.

2. Bionomic label. This second label may have additional information, such as the host plant, collecting method or habitat (this can sometimes be added to the first label if there is room).
3. Identification label. As well as the name of the species, identification labels should give the name of the identifier and date of identification. They should not be removed or discarded, even if another authority adds a label with a different name at a later date. The earlier label could still be important, especially if the specimen concerned has been referred to in the scientific literature.
4. Curatorial labels (for example, showing accession details or type status).

Labels must last as long as the specimens and many years may pass before faults begin to appear. Therefore, inks and label paper or card must be of archival standard (see Appendix II, Papers, inks and label conservation). A minimum paper weight of 100 gsm (grains per square metre) is recommended for both dry and wet collection labels.

*Pinned specimens*

Pinned specimen labels should be kept as small as possible and not greatly exceed the area taken up by the specimen. An ideal size is 12 X 8 mm and several labels below each other on the pin are quite acceptable. It is usual for the locality label to be uppermost, nearest to the specimen, with the identification label added last. Additional information can go on the underside of a label but many workers find this unacceptable as it is easily overlooked. Labels should always be orientated under specimens in a standard way for easy scanning. Some museums now use bar codes on their labels (see Appendix I on Documentation). Usually the label reads from left to right when the pin is held in the right hand. Try to avoid the pin going through any print. Pinning blocks with holes of several depths may help to standardize heights of labels but make sure that the points of the pins are not damaged in the block.

*Microscope slide labels*

Care must be taken when selecting slide labels. Some commercially available slide labels are unsuitable for permanent preparations because of poor quality paper or glue. For example, glues with a natural rubber base deteriorate, especially when exposed to light or heat. If a self-adhesive label is used, ensure that it is of archival quality (e.g. foil-backed, with acid-free adhesive). Paper and glue must also be able to withstand heating in a drying oven. Some institutions use thick card (i.e. Bristol board) as slide labels. These labels are thicker than an average specimen mount and are glued at each end of the slide with neutral pH PVA adhesive. This system serves to protect the mount when the slides are stacked on top of one another or stored vertically in a cabinet.

The accepted format for slide labels is to write the specimen data on the right-hand label and the identification on the left. It is also important to include details of the mountant used, date of preparation and the preparators name. To avoid loss of data if labels become accidentally detached, a reference number should he scratched into the surface of each slide with a diamond pencil.

*Curatorial points relating to specimen labels*

Colour coding of labels of type specimens makes their retrieval from collections easier. Red is widely used for primary types (particularly holotypes) but there is no accepted convention. Microscope slides may have a coloured line on the label or the identification highlighted in colour. In some old collections a piece of coloured paper may be the only indication that a specimen is a type. Colour coding can be used for other purposes, for example to draw attention to reared specimens, but too many colour codes can lead to confusion. If a colour coding system is to be used it is important to use inks that will not fade (see Appendix II on papers, inks and label conservation).

*Cross-reference labelling*

Parts of specimens may be detached and made into separate preparations, for instance, slides of genitalia or fragments mounted on scanning electron microscope stubs. Such preparations are difficult to keep with pinned or ethanol-preserved specimens. Also there may be, for example, associated larvae or host specimens

which need to be preserved by a different method. To overcome the possibility of disassociation, there must be adequate cross-referencing on the labels of all such material. We know of no uniformly applicable system that we can recommend and all the examples we are familiar with have good and bad points. However, with the advent of electronic databasing systems, cross-referencing can become easier, provided that all separate parts of an individual specimen are given a common identifying number.

*Code numbers*

Although the practice is discouraged, in the past it has been fashionable to attach only a number to a specimen and to record all details in an index (on cards or in a book). At worst such indexes are destroyed and the specimens lose most of their value. At best the information can be transcribed accurately on to specimen labels at a later date. Other problems arise when several `numbered' collections get mixed up, and sometimes indexes thought to be lost turn up. Code numbers are sometimes added to specimens in addition to data on labels. These may relate to more extensive information entered in a field notebook together with the collection details. Such notebooks should be carefully preserved. The use of bar codes in insect collections is still open to debate and further information can be found in Appendix I on documentation.

*Provenance of specimens*

Labels often provide the only clues to the origin and history of a specimen because collectors and curators are sometimes lax in documentation and record keeping. No matter how tattered or indecipherable they might appear, old labels can be important in tracing more information about a specimen, leading to the recognition of a type, for example. An important reference work is Horn *et al.* (1990), a compendium of (amongst other things) handwriting on specimen labels. Institutions which hold historical collections should keep an archive of label handwriting for future reference.

*Accession labels*

In many institutions it is normal practice to integrate material from different collections into a single system. When this is done it is of great importance to attach to each specimen an accession label to indicate its historical origin. If the institution operates a numbered register system for keeping a record of collections, the accession label should bear the registration number. The specimen register can provide an important additional source of data (see Appendix I on documentation and Chapter 9 on policies and procedures).

***Sending material through the post***

It is often necessary to send specimens through the post in order to make material available to a wider scientific community. However, this should only be clone if it is possible without unacceptable levels of risk to the specimens. Good packing can ensure that specimens are subjected to minimum levels of physical stress during transit but material should not be dispatched to countries with unreliable postal systems (see Chapter 9 on policies and procedures). Insect specimens should be pinned into strong postal boxes lined with a material such as Plastazote that will grip the pins adequately. In the case of insects with long or *heavy* bodies, it may be necessary to cross-pin these to prevent them from swivelling. Never enclose insecticide crystals, moth-balls or similar substances with specimens as they may lead to severe damage. The postal box should be sealed to prevent the entry of dust and pests and enclosed inside a much larger outer box, cushioned all round with a thick layer of shock-absorbing packing material (Fig. 2.14), (Peigler, 1992; Walker and Crosby, 1988). Some fragile insects such as dragonflies are best sent unset and papered hut still packed in the same way. Very fragile or unique specimens should not be sent through the postal system. It is sometimes possible to have specimens hand-carried by a colleague but the same care must be given to packing and protecting such specimens.

## Organization and storage of collections

The basic rules of storage are that specimens must be kept in total darkness and excessive heat and humidity avoided (see Chapter 7 on the collection environment), but guidance on



**Figure 2.14** A packed parcel in cross-section. Note the amount of polystyrene packing surrounding the specimen box; the packing must fill all the space but not be compressed too tightly — it acts as a cushion to absorb physical shocks in transit.

organization is less easy. Most insect collections are not arranged arbitrarily or alphabetically; they are organized taxonomically, following some accepted classification. Most institutions possess several distinct collections, but exact arrangements will vary according to a number of factors. Pinned, fluid preserved and microscope slide collections are almost always kept separately. Other factors which affect curatorial policy (and the ability to implement it) include size of collection, its purpose, level of use, availability of resources (including expertise) and historical significance. A large museum may aim to unify most material in a single large collection, but keep separate collections of the local fauna, common pests and type specimens. All of these matters are for local decision. For example, it is debatable whether or not type specimens should be kept separately. Most institutions prefer to keep their type collection separate, thus making the specimens more accessible if they need to be removed quickly in case of emergency. Moreover, they are less likely to be damaged if kept away from the more frequently handled reference series material. On the other hand, types are not always easy to identify and cannot be segregated easily.

Also, a pest infestation in a separate type collection can lead to catastrophic losses.

***Pinned material***

Pinned insects are traditionally stored either in cabinets with shallow drawers or insect `store-boxes'. In both containers a soft pinning base is necessary to grip the tips of the pins and hold them securely. In the past, cork, balsa-wood, peat and expanded polystyrene have been used, but the most acceptable material currently available is white cross-linked polyethylene foam, available under the trade name Plastazote. The recommended grade of Plastazote is 4.05 kg per cubic metre, in sheets 9–12 mm thick. Unlike cork, for example, it is white and therefore does not need covering with paper (although it becomes statically charged and may get very dirty if left exposed). Cutting the foam at a slight angle, with the upper surface a fraction larger than the container, will ensure a snug fit in the box or drawer. Conservation grade polyvinyl acetate (PVA)-based adhesives can be used to hold it in place; but in some situations old-fashioned gelatin-based glues may be better. Avoid using contact glues as they may not penetrate into the foam sufficiently or may react with other materials to release harmful gases. Moreover, if a fumigant is present, the reaction between the fumigant and the contact glue may not allow the cement to harden. When fitting any pinning base, ensure that it fits snugly around the perimeter, as it is extremely difficult to retrieve specimens which fall down the sides. A gap also encourages dermestid pests. If Plastazote is not available, cork covered with lining paper is an alternative but ensure that the cork, lining paper and glue are of archival quality. Ensure that the adhesive is not hygroscopic — damp paper and/or glue can react with the pins so that they become permanently corroded in place or even break off in extreme cases. One reason for persisting with cork is that good quality, old drawers are often too shallow to fit with Plastazote and still leave room for specimens and pins.

Dried insects are fragile. Lift the lids off drawers and open store-boxes very slowly or else an inrush of air may blow off delicate legs, antennae and wings. Also make sure that one-sided store-boxes are' opened with the base flat so that any loose specimens will



**Figure 2.15** An insect cabinet with the door open. The cabinet is metal but the drawers are wooden. Note the foam seal around the door.

remain in the box rather than falling out. Do not slam drawers shut; pull them out gently. When placing boxes on shelves, do so carefully — always remember the fragile contents. Never let glass lids rest on pin heads as the resulting movement can cause damage to the specimens.

*Drawers and unit trays*

Drawers and cabinets can be constructed from wood or metal and there are a number of kinds available commercially but, at least in Europe, there has been relatively little attempt to standardize sizes and constructional details. For a discussion of materials used see Chapter 7 on the collection environment. A modern cabinet (Fig. 2.15) will usually hold up to twenty drawers although some older cabinets house forty or more. Within cabinets, drawers should be interchangeable and cabinet doors should be seated on a flexible seal. Open racks of drawers are not recommended as specimens are vulnerable to penetration of light, dust and pests.

Drawers should not be too large or heavy. The wood-framed glass lids must fit well to keep out pests. Other considerations include the need to carry the drawer from cabinet to work-bench and the ability to manipulate it and reach specimens easily when it is on the bench at the side of a microscope. Townes (1973) covers most of the practical points involved. An ideal size of drawer is about 450 mm square and 60 mm deep (outside dimensions).

The most practical system for the storage of small specimens is based on `unit' trays or 'pinning' trays. The trays are of various



**Figure 2.16** A cabinet drawer illustrating three different sizes of unit trays. The glass lid has been removed.

standard sizes, multiples of one another, so that they can be moved around and rearranged in the drawers with minimum handling of the specimens themselves (Fig. 2.16). The unit trays are best made of sturdy white, acid-free cardboard, glued with conservation grade adhesive and lined on the base with Plastazote. Some museums use plastic trays. The unit tray system is not necessarily the best for all groups of insects (for example, some butterflies are so large that the drawer becomes the equivalent of a unit tray). An alternative system is to use Plastazote or corklined strips which can be moved and rearranged in a similar way to unit trays. The drawback to the strip system is that it is easy to pin specimens too close to the edges, where they are very easily damaged when strips are moved.

New cabinets are very expensive but it is worth bearing in mind the potential value of old cabinets. They may not provide the best housing for an insect collection but they may fetch high prices as pieces of `antique' furniture, and thereby help finance the purchase of new cabinets. However, sale of cabinets may raise legal and ethical problems in some museums. In the case of early `historical' collections, the cabinet is an integral part of the collection itself, but very few early collections remain in their original cabinets. If specimens must be removed for their protection, then the original cabinets, drawers and specimen arrangements should be fully photographed and documented. The use of old cabinets to house museum collections presents the following problems: non-interchangeable drawers; shrinkage of bases causing cracks which may admit pests; and worn runners and warping sides causing drawers to stick and resulting in jarring damage to specimens (this can sometimes be alleviated by applying beeswax or paraffin wax to the runners).

### *Store-boxes*

Insect store-boxes (Fig. 2.17) are sometimes a practical, if not ideal, alternative to cabinets. There are various kinds available and careful consideration should be given to requirements before opting for a particular variety. Large, double-sided boxes are generally best avoided; not only are they cumbersome and heavy, but pins on opposite sides often become entangled when the box is closed and so damage the specimens. The boxes should be kept in pest-proof cupboards if at all possible, and not stored on open shelves. The great disadvantage is that boxes must be opened before specimens can be examined or checked for pest attack. A well arranged, fully documented system will reduce unnecessary handling.

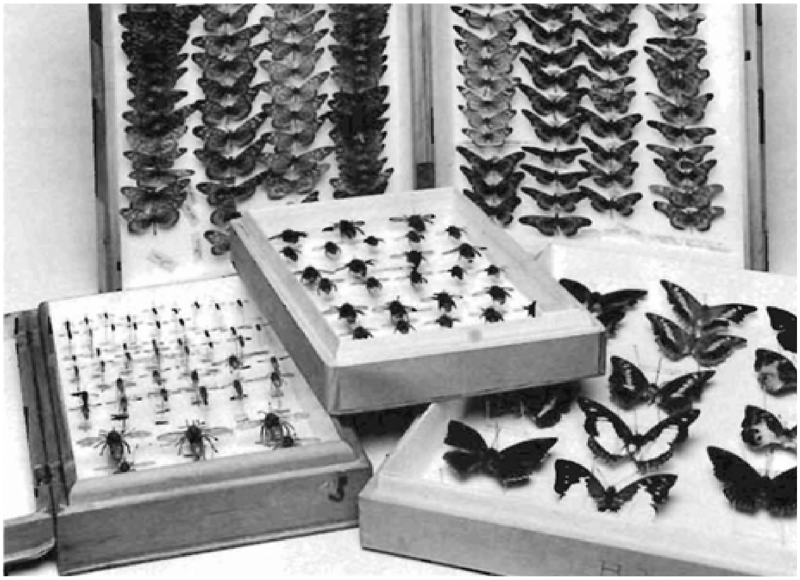


**Figure 2.17** Various designs of store-boxes, either hinged or with a removable lid. The store-box in the background is doubled sided, a design not generally recommended, but in this example both leaves are very deep and thus there is no possibility of pin heads touching opposite specimens when the box is closed.

### *Storage of papered specimens*

Because of their extremely delicate nature, Odonata collections are frequently kept in paper triangles or envelopes and these are most frequently stored in filing cabinets or cardboard boxes. Many museums also hold collections of other `unset' insect specimens in the original envelopes in which they were collected in the field. It is important to ensure that the individual paper packages are not packed too tightly together or damage will ensue. A well organized, fully databased system will improve access and reduce handling to the minimum. Care should be taken to exclude pests and dust and it is best to enclose files and boxes of specimens within cabinets with pest-proof doors.

### *Microscope slides*

For economy of space, slides with relatively thin mounts can be stored vertically, in purpose-made cabinet drawers. Shallow-drawer, metal office cabinets can be modified easily and will provide adequate conditions for slides (Fig. 2.18) provided that the collection's environment is relatively free of dust (such cabinets do not usually protect slides from dust and dirt). With vertical storage, it is important to ensure that the slides do not touch or rub against each other. This problem can be avoided by using thick card labels each side of the coverslip, which serve as a buffer as well as a surface for data. The slides can be protected further by a polyester sleeve.

Many mountants take a long time to harden and the specimen will slide down the mount if it is stored vertically too soon after preparation. In fact, all slide mounts are to some extent `fluid' and, where possible, flat storage is recommended, particularly in the case of very thick preparations which never harden completely. Flat storage of microscope slides has the advantage that every slide is immediately viewable, but collections take up more space. Purpose-built slide trays and cabinets are available for flat storage (Fig. 2.19). An alternative and cheaper method of flat storage is to use slotted slide boxes which can then be stored upright like books on shelves (thus keeping the slides horizontal) (Fig. 2.20). However, great care must be taken that such boxes are marked so that they are opened the correct way up, otherwise slides may fall out and break.

Apart from the danger of breakage, glass microscope slides (once fully dry) are relatively robust and require no special handling or storage conditions, although they should be kept in total darkness and as cool as practicable. Consideration should be given to the great weight of large slide collections when choosing a place to store them.




**Figure 2.18** Vertical storage of microscope slides using a modified, shallow-drawer, metal office cabinet.

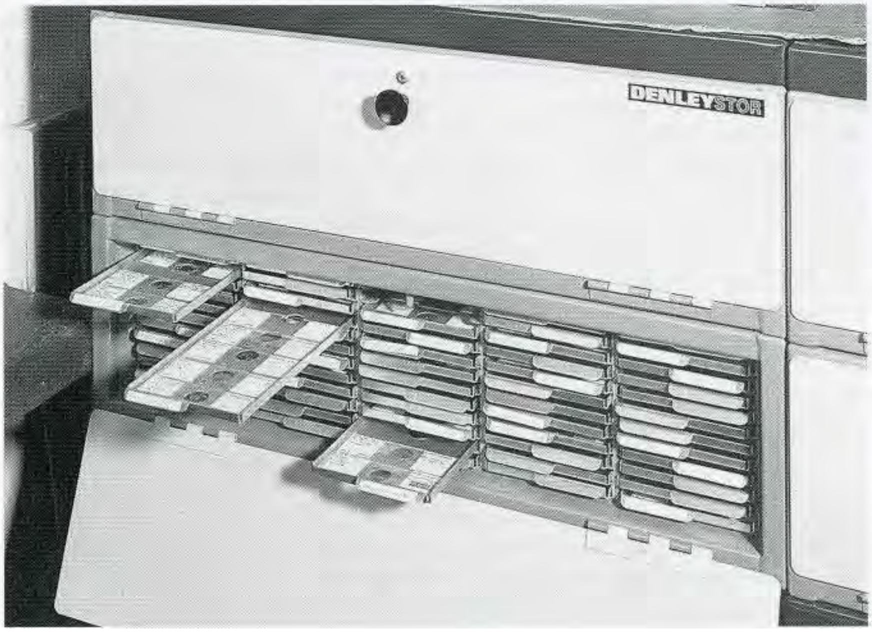



**Figure 2.19** Purpose-built flat storage for microscope slides.

***Scanning electron microscopy (SEM) mounts***

Specimens prepared for examination by scanning electron microscopy are mounted on aluminium discs called `stubs'. For optimum results, specimens are coated with a microscopic layer of metal, but it is possible in some cases to obtain acceptable results without coating. Although it is possible in some cases to remove metal coatings after examination (Golden, 1989), coating should be considered as potentially irreversible and therefore a



**Figure 2.20** Purpose-built slotted slide box (which can be stored upright on shelves.

destructive process. It should be avoided for irreplaceable specimens such as types.

After examination, particularly of uncoated material, specimens can be removed from the stubs and returned to the collection. To facilitate this, specimens prepared for SEM study can first be attached to glass coverslips which are then mounted on SEM stubs. The coverslips can later be removed and mounted on microscope slides.

In many cases, particularly where material may be needed for further SEM study, it is more practical to store coated specimens mounted on their stubs. Golden (1992) illustrates a polystyrene box custom-made to house them, but they can also be stored in shallow polystyrene boxes lined with Plastazote which has been punched with small holes to hold the pegs of the stubs. Overcrowding in boxes should be avoided as it is easy to damage other specimens when removing and replacing stubs. Some metal coatings will last well but will inevitably deteriorate with age. Where necessary, it is sometimes possible to recoat such specimens.

### *Unsorted, unprepared material*

Many museums have large accumulations of unsorted, unprepared material and if it is of value it must be properly cared for.

*Dry storage*

Insects which are collected `dry' in the field are often temporarily stored in large numbers between layers of tissue packed in boxes, or single insects such as butterflies are stored with their wings folded together in paper triangles, within boxes or tins (see above). Material stored in such a fashion is especially vulnerable to insect pest attack and must be checked frequently. Ideally layering boxes should be stored in sealed containers or plastic bags, but ensure that the specimens are perfectly dry first and add a few crystals of thymol to prevent growth of mould. Microlepidoptera can be stored temporarily (after micro-pinning and before setting) in shallow polystyrene pinning boxes lined with Plastazote.

*Deep-freeze storage*

Freshly caught specimens which are to be pinned can be sealed in field layering boxes or other suitable containers and stored for short periods in a deep-freeze. Specimens should not be allowed to dry out before placing them in the deep-freeze and cardboard containers should be placed in plastic bags and sealed to prevent this. When removing material from the freezer, allow it to reach room temperature before opening the container, as this will prevent wetting due to condensation.

### *Fluid collections*

General information on storage of fluid-preserved material is provided in Chapter 5 on fluid preservation, but insects and other small arthropods have special requirements which are described here.

Specimens are best stored in straight-sided, flat-bottomed glass vials with a polyethylene cap or a plug of cotton wool (Fig. 2.21). Avoid cork, rubber or neoprene stoppers, as they will eventually degrade or leach substances into the preservative (Plates 9 and 10). The practice of plugging vials with cotton wool is not recommended for some adult insects, as tarsal claws may become entangled, leading to difficulties when removing specimens. Vials

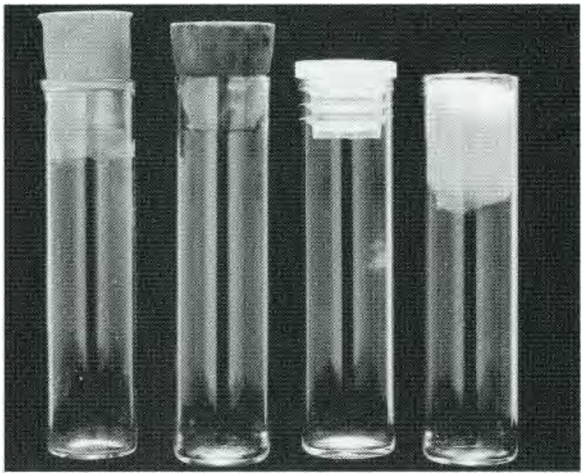


**Figure 2.21** A selection of flat-bottomed glass vials illustrating different stoppers. Left to right: rubber, cork, polyethylene and cotton wool Rubber and cork stoppers are not recommended.

should not be packed too tightly with specimens, as wings, legs and antennae will crumble and break. Individual vials should contain only specimens collected together.

The vials are filled as completely as possible with preservative and care should be taken to equalize pressure inside and outside the vial when inserting polyethylene caps, otherwise they may pop out again. Vials are best stored in jars of preservative, lined on the bottom with a thin layer of cotton wool or other conservation grade wadding to protect them. This reduces the risk of vials drying out and simplifies topping up. Vials plugged with cotton wool should be inverted into the jars but those with polypropylene caps should be stored upright; otherwise, if a cap should pop off, specimens will quickly disperse in the preservative. For economy of space and ease of handling, storage jars should have wide mouths and straight sides (see figures in Chapter 5 on fluid preservation).

### *Documentation and indexes*

Catalogues and indexes are needed for easy access to specimens in a collection, for the retrieval of information, and for record purposes. However, collections should be self-indexing as far as possible to minimize the work of making indexes and keeping them up to date. A lot of indexes merely duplicate information intrinsic to the specimens and labels or the arrangement of the collection. Simple yet effective ways of finding ones way around the collection are best. With a systematically arranged collection the first requirement for easy access is to code each cabinet and drawer, box or jar. Then an index can be compiled to the taxa. It is often not necessary to go below genera as most genera will have only a limited number of species. The data are best held on a computer, as a simple word-processed file for a small collection or in a database for larger and more sophisticated arrangements. A print-out is often convenient for day-to-day use and for recording changes for transfer to the computer system. For further information, see Appendix I on documentation.

## Remedial conservation

In the right conditions dead, dry insects and arachnids remain stable more or less indefinitely, while specimens in alcohol or on microscope slides may show signs of gradual deterioration. However, we are not aware of any definitive work on the long-term deterioration of these sorts of biological materials. The majority of conservation problems will relate to inadequate curation or application of inappropriate techniques, rather than to changes in the specimens themselves. For pinned collections the most frequently needed general conservation measure is to transfer the specimens to safer storage. Most specimens will then become stable and are probably best left alone until specialist advice and help is available. Conservation action may be appropriate in a limited way only (see also Chapter 9 on policies and procedures). Questions which should be asked before any remedial action is undertaken include:

- What is the value of the material in comparison to the cost of conservation measures?
- Has the damage limited its scientific value?
- Is further damage likely and is it preventable?
- Is the necessary expertise available?

Often specimens become very dusty if they have been left uncovered or stored in badly sealed containers for any length of time, or they may be covered in moth scales from collection in a light trap. Before they can be examined they may need to be cleaned. The specimens will not usually deteriorate if left in a dusty condition as long as they are transferred to better storage containers.

Individual specimens can be cleaned as already described, using brushes or ultrasonic cleaners. For very delicate specimens, fine cleaning can be achieved using vacuum 'micro-forceps'. The relatively new technique of laser cleaning is still being refined but has considerable potential for use on insect specimens. Current drawbacks are that the process is both time-consuming and costly.

### Labels

It is very important when conserving specimens to ensure that old labels are retained (often the original handwriting is the only clue to the origin of an important specimen and can be vital for tracing types). Trying to reinterpret label data can often give rise to errors. Old specimen labels damaged by successive repinning or paper deterioration should be glued carefully, with starch paste, on to a small piece of fresh label paper as a support. Poor quality labels which have become brown and brittle can first be de-acidified in a 1% solution of barium hydroxide in methanol (see Appendix II on papers, inks and label conservation).

### Replacing pins and double mounting old specimens

Old collections were usually pinned with non-stainless pins and often become weak or corroded. Various methods for removing pins have been discussed in the literature, including use of Barber's fluid (May, 1958) or employing an electrical current to heat the pin (Upton, 1991), but this latter method has been found to be unreliable unless used with great care. In most instances, simple methods and patience are adequate.

With large pinned specimens such as grasshoppers or beetles, remove the labels and relax the pinned specimen by soaking it in a strong solution of one part colourless liquid detergent to three parts warm water or by placing it in a relaxing chamber until soft (it is not recommended to soak hairy insects such as bees or moths, or specimens required for DNA studies). Gently ease the pin out, twisting the pin to assist its removal. Use watchmakers forceps as shown in Fig. 2.2. Then carefully repin the specimen using a stainless steel pin while it is still relaxed. If the old pinhole is too large for the replacement pin, place a drop of water-soluble glue on the pin shaft slightly below where the specimen will rest, before moving it up over the glue. Some specimens can be softened sufficiently to remove their pins by applying a few drops of methyl alcohol to the thorax, but this must be carried out with care and is not appropriate for very delicate insects.

Small specimens are not so robust and, if possible, it is best to leave the old pin in the insect and pin it to a double mount (see Fig. 2.8). If the old pin is too long, use a pair of fine wire-cutters (piano wire-cutters are ideal) to trim it to a length suitable for double mounting. This technique does, however, require great care as violent jarring by wire-cutters can easily damage a delicate specimen, even when it is relaxed. Hold the pin firmly in the forceps between the specimen and the cutters, to lessen the shock, then slowly apply increasing pressure to the cutters. If the old pin has broken just underne2'h the insect, the only solution may be to glue it directly on to the double mount.

### Soaking specimens off cards

Specimens are sometimes glued flat on to pieces of card which makes examination of the ventral surface almost impossible. These specimens can easily be removed for study by soaking them off the card using a few drops of warm water around the specimen (before discarding the old mount, make sure the card does not have label data on the underside, or legs etc. attached). If the glue is not watersoluble, then ethanol, acetone or ether may act as solvents.

***Removing verdigris***
Verdigris is a considerable problem with insects direct-pinned with brass pins. It is a green crystalline deposit of esters formed as the result of a reaction between the fatty acids of the insect's body and the copper content of the pin. Particularly in conditions of high temperature and relative humidity, verdigris can develop rapidly and will eventually destroy the specimen. Insects most at risk seem to be large beetles and moths but other orders are also affected. The only satisfactory solution is to remove the corroded pin and replace it with a new stainless steel one after all traces of verdigris have been removed from the specimen by careful brushing. In extreme cases, specimens may begin to disintegrate and must either be glued on to card or stored in gelatin capsules. For less seriously affected specimens it is possible to clean off verdigris without removing the pin by brushing acetone on to the affected area, allowing it to soak in and then gently breaking away the verdigris with a fine brush (Walker and Crosby, 1988). This process will probably need to be repeated at regular intervals, so pin replacement is generally the better option. As a precaution, clean specimens in a fume cupboard or well ventilated room when using acetone or other volatile solvents.

***Restoring shrivelled and dirty specimens*** For restoring specimens which have become shrivelled and will later be stored in ethanol, or large, dry specimens (for example grasshoppers) needing repinning or cleaning, the following technique is useful:

1. Remove labels and soak the specimen in one part colourless household detergent to three parts warm water for no longer than fifteen minutes. These proportions are only a guide, as much depends on the detergent and the state of the specimen.
2. Gently agitate the container occasionally. If the specimen is not sufficiently relaxed in the detergent solution after fifteen minutes, remove it from the solution and place in a relaxing chamber for up to two hours. This should be sufficient time to soften it completely.
3. If the specimen is to be pinned, rinse in acetone, arrange legs, and pin immediately.
4. If the specimen is to be preserved in ethanol, rinse in ethanol before storing.
5. An alternative method is to use a 1% solution of trisodium phosphate ($Na_3PO_4$) in cold water. Sink the specimen with a drop of detergent, leave for 24 hours, and then wash it in ethanol.

A detailed description of a restoration technique applied to bumble-bees is given by Milliron (1971). Upton (1991) recommends soaking specimens in Decon 90 for sixteen hours, then thoroughly rinsing and immersing in water until restoration is complete.

***Restoration of specimens on slides***
The mounting medium used to preserve specimens can dry out and shrink, causing the appearance of scattered fragments of mountant under the coverslip. In severe cases this can damage the specimen but usually the specimen can be retrieved. Shrinking usually occurs with water-based mountants such as 'Hoyers' when the coverslip has not been ring -sealed to prevent desiccation. With patience and care specimens can be restored, depending on their age. However, this process is a very delicate one and restoration should be done by a specialist who is familiar with the group. The basic technique is as follows:

1. Pipette a drop of warm, sterile water under the coverslip and leave for a few minutes until the water has been absorbed by the mountant. Repeat the process until one can gently ease the coverslip off the slide.
2. If water has no effect, try xylene, as the mountant may be resin-based (e.g. Canada balsam).
3. Finally, if the mountant will not shift, it may be Euparal (which has its own solvent).
4. After this process, locate the specimen either on the slide or the coverslip using a dissecting microscope.

Ensure that the above sequence of solvents is followed as the wrong solvent can cause irreparable damage to the specimen. Usually it is impossible to reorientate specimens from the original mounting position, but the specimens can often be remounted satisfactorily using a standard mounting technique for the

group concerned. Brown (1997) indicates that blackening `Berlese' mountant can be dissolved in acetone and some specimens can be remounted successfully, but in the case of aphids the insect cuticle is still irrevocably damaged'. Fain (1980) gives a procedure for remounting mites in water-based mountants without damaging the specimens. Ensure that all microscope slides in a collection which have a water-based mountant are ring-sealed with Euparal follow ed (after it has hardened) by Murrayite. For fluid mounts see Chapter 5 on fluid preservation.

## Other invertebrates

Although other groups of invertebrates are mostly stored in alcohol or other fluid preservatives (see Chapter 5 on fluid preservation), some Mollusca, Bryozoa. Porifera, Coelenterata (corals) and Echinoderms are preserved dry and some old dried specimens are of considerable historic importance. Dry specimens have traditionally been stored in glass-topped cardboard or wooden 'museum boxes' with specimens supported by cotton wool or acidfree cellulose wadding. The boxes are either housed in drawers or on shelving, sometimes within cabinets but often stored openly. Storage in glass-topped boxes makes viewing easy but leaves specimens vulnerable to fading of contents unless they are kept in darkness. Mathias (1994) suggests that a cheaper way to store this material is to rest specimens on cellulose wadding or acid-free tissue in acidfree card trays, enclosing them in self-sealing polythene bags but, as Mathias pointed out, a major disadvantage here is the waste of space as trays cannot be packed close together.

Small specimens are often preserved in glass vials. Burns and Ballantyne (1992) describe a system for keeping small specimens in gelatin capsules within glass vials. These are stored in cabinet drawers lined with polyethylene foam, drilled with holes to accept individual vials.

### Mollusca

In modern shell collections, individual specimens are often enclosed in polythene bags and stored in open unit trays (Plate 8). Mathias (1994) recommends storage of small molluscs in gelatin capsules enclosed within glass vials packed at either end with wadding. Taylor (unpublished) of Hampshire County Council Museums Service recommends storing small specimens in snap-top polystyrene boxes lined with polyester wadding to cushion specimens. These boxes are stored in stove-enamelled steel cabinets. Garback and Spamer (1992) recommend similar boxes, lined on the base and sides with polyethylene foam and stored in drawers subdivided by acid-free hoard strips. The National Museum of Wales uses similar boxes stored in open fibreboard trays in a compactor system. The use of self-sealing polythene bags for storage of larger shells is widely adopted in museums today.

A major threat to shell collections is 'Byne's disease' (Plate 21) — not a disease at all but an efflorescence resulting from reactions with organic acid fumes released from wood and other materials used to make storage cabinets (Shelton, 1996). Tennent and Baird (1985) observed efflorescences to be mostly calcium acetate or calcium acetate—formate double salts. All affected specimens had been stored long-term in oak cabinets (choice of materials for cabinet and drawer construction is discussed in Chapter 7 on the collection environment). Cotton, cork and non-archival plastics should also be avoided, and labels and tags should be on acid-free paper. Da Fonseca and Thome (1992) recommend washing affected shells with fresh or distilled water hut the general opinion seems to be that washing should be avoided if possible. Moreover, Shelton (1996) draws attention to the fact that humidity changes can split thin shells. The efflorescence can sometimes be carefully brushed away but it is recommended that this is only done by an expert and the best practice is to transfer the specimens to a more suitable environment.

Protective coatings on shells have been tried but are not recommended. Coatings based on shellac, cellulose nitrate and polyvinyl alcohol tend to shrink and yellow over the years, sometimes causing actual damage and also tending to act as dust traps, obscuring the surface structure.

Shells should ideally be stored in archival boxes and trays housed in steel cabinets with powder-paint finishes. However, this is not always practical, particularly in the case of

large collections or where funding for new cabinets is not available. Existing drawers can be coated with acrylic latex paint containing calcium carbonate to prevent off-gassing but this is also expensive. Storage of specimens in sealed polythene bags (ideally, anoxic enclosures; see Burke, 1996) provides a relatively cheap and simple answer as it effectively isolates the specimens from acid fumes.

### Bryozoa

At The Natural History Museum, London, specimens are traditionally stored in card boxes in subdivided wooden trays housed in cabinets. The current method is to store specimens in polystyrene boxes similar to those used for mollusc shells. Sometimes several specimens are stored in larger boxes, layered between sheets of acid-free tissue. For larger specimens, the boxes are lined with Plastazote to provide cushioning. Compactor storage is not recommended for such bryozoans as they are too fragile. Some older material is mounted on herbarium sheets but can be vulnerable to damage due to pressure exerted by the layers of sheets. It is proposed to remount the specimens in framed mounts to provide some protection from crushing (Spencer Jones, pers. comm.).

Small specimens are often slide mounted. Old slides are dry mounts with specimens glued on to a wide range of different slides — some early slides are made of wood or metal. Current procedure is to mount specimens in Canada balsam on glass slides.

### Porifera

Glass-topped boxes have been traditionally used to store dry specimens and are still used today. Ideally they should be lined with acid-free tissue, Plastazote or some other conservation grade cushioning material to provide adequate protection to specimens. Large specimens that are too big to go into cabinets can be protected by covering with a Tyvek dust-sheet.

### Coelenterates

Dry-preserved corals and gorgonians have traditionally been stored on shelves or in drawers in wooden cabinets and it is only recently that curators have taken the step of enclosing specimens in polythene bags or covering large specimens to prevent dust contamination. Coral quickly becomes dirty and discoloured, although commercial suppliers often bleach their specimens and sometimes retouch them with colours – practices that are not recommended for scientific specimens. Halsey (pers comm.) has experimented with cleaning coral by steeping it in water with a wetting agent (Teepol) for several hours and then air-drying it at ambient temperature. Results seem to be excellent, but this method has yet to withstand the test of time.

### Echinoderms

Dry specimens are traditionally stored in glass-topped boxes similar to those used for mollusc shells. Fragile specimens are protected from damage by careful cushioning with cotton wool or acid-free tissue.

## Public display and teaching collections

### Display

Because of the extremely fragile nature of most invertebrate specimens, great care must be taken when selecting specimens for display. Valuable material, for instance types and voucher specimens, should not be put at risk by use in displays. No matter how much care is taken there will inevitably be a higher risk of damage to display specimens (see the story of Lister's Conch in the Preface, p. xv. Select large, robust specimens which will be easier for the public to see and less prone to damage.

Another problem facing curators is that the design of displays for the public does not always take conservation issues into account. Invertebrate specimens displayed in dioramas or large open areas are very difficult to protect from dust or pest attack. On the other hand, specimens displayed in closed cases can be protected to a much greater degree, even to the extent of enclosing insecticides to prevent pest attack. Displays must be kept away from

direct sunlight and lights should have UV filters fitted over them. For further details see Chapter 7 on the collection environment.

For small temporary displays, various glass-topped wooden or cardboard boxes are available commercially, but it is possible to modify a store-box by inserting a glass panel in the lid. Glass-topped cabinet drawers can also be used. Liverpool Museum has special display cases designed to exhibit drawers from their main collections and displays are rotated on a regular basis.

Freeze-dried specimens are excellent for display purposes as they retain their colour and are less susceptible to shrinkage (see p. 43).

### Teaching collections and `hands on' material

Invertebrates are generally too fragile to be handled by the public, although it may be possible to provide robust shells, coral fragments, insect-bored wood and similar items for children to touch. Such material should not be taken from reference or study collections unless it is of poor quality and without data. It is sometimes possible to mount robust specimens securely within sealed plastic boxes for handling, although these may have a relatively short life.

### Embedding insects in plastic

Small invertebrates such as insects and their larvae can be very satisfactorily embedded in clear plastic resin, although this has the disadvantage that it is an irreversible technique, and the plastic tends to deteriorate and discolour after a few years of use (Plate 22) (see p. 107 for repairing damaged plastic surfaces). Nevertheless, it is an excellent method for a `hands on' display where school-children can handle specimens and still see all the features, while the specimens are fully protected. Bergmans (1968) and Moulds (1975) describe techniques for embedding insects in resin.

### Microscopes

Small specimens such as insects are often difficult to display but many museums are now using video cameras mounted on fixed-focus microscopes for viewing exhibited specimens. Some museums supply specimens in transparent boxes that can be viewed under such microscopes. Ideally these activities should be closely supervised by a member of staff.

## Acknowledgements

The authors wish to gratefully acknowledge the help and advice given by colleagues at the CAB International Institute of Entomology and The Natural History Museum, London. We would also like to thank Philip Hurst (Photographic Unit of The Natural History Museum) and Julie Harvey (Entomology Librarian, The Natural History Museum, London). Murry Upton (Australian National Insect Collection) is also thanked for his kind advice. We would also like to acknowledge permission from the Department of Scientific and Industrial Research, New Zealand for the use of figures 2-5, 29 and 34 from Walker and Crosby (1988) and the CAB International Institute of Entomology for the use of figure 2.14 from Instructions for Users of the Identification Service (1989).

## References

Bergmans, J. (1968). Embedding entomological specimens in an improved polyester resin. *New Zealand Entomologist*, **4**, 61-66.

Brown, B.V. (1990). Using Peldri II as an alternative to critical point drying for small flies. *Fly Times*, **4**, *6*.

Brown, B.V. (1993). A further chemical alternative to critical-point-drying for preparing small (or large) flies. *Fly Times*, **7**, 10.

Brown, B.V. (1995). A further chemical alternative to critical-point-drying for preparing small (or large) flies. *Chalcid Forum*, **18**, 5.

Brown, P.A. (1997). A review of techniques used in the preparation, curation and conservation of microscope slides at The Natural History Museum, London. *The Biology Curator*, **10** (Supplement). 1-33.

Burke, J. (1996). Anoxic microenvironments: a simple guide. *SPA/IC Leaflets*, **1**, *1-4*.

Burns, J. and Ballantyne, M.R. (1992). System for storing small specimens. In *Storage of Natural History Collections: Ideas and Practical Solutions*. (C.L. Rose and A.R. de Torres. eds), pp. 209-210. Society for the Preservation of Natural History Collections. Pittsburgh.

Carter. D.J., Vogler, A.P. and Vane-Wright, R.I. (1997). Notes on basic collecting techniques for morphological and molecular studies of Lepidoptera. *Metamorphosis*, **8**, 99-106.

Cowan, D.P. (1995). Another method of drying chalcidoids. *Chalcid Forum*, **18**, *4-5*.

Craig, D.A. and Craig, R.E.G. (1987). Simuliidae (Diptera: Culicimorpha) of Rarotonga, Cook Islands, South Pacific. *New Zealand Journal of Zoology*, **13**, 357—366.

da Fonseca, A.L.M. and Thorne, J.W. (1992). Degradation of seashells in dry collections — the Byne disease' or efflorescence. *Siratus*, **13**, 14—15.

Deitz, L.L. (1979). Curatorial note on genitalia vials. *New Zealand Entomologist*, **7**, 10— 11.

Fain, A. (1980). A method of remounting old preparations of acarines without raising or displacing the cover slip. *International Journal of Acarology*, **6**, 169—170.

Garback, M.A. and Spamer, E.E. (1992). Boxes for dry shell collections. In *Storage of Natural History Collections: Ideas and Practical Solutions*. (C.L. Rose and A.R. de Torres, eds) pp. 185—186, Society for the Preservation of Natural History Collections, Pittsburgh.

Gauld, I. D. and Bolton, B. (eds) (1988). *The Hymenoptera*. British Museum (Natural History) and Oxford University Press, London.

Gilbert, P. and Hamilton, C.J. (1990). *Entomology: A Guide to Information Sources*, 2nd edn. Mansell, London.

Golden, J. (1989). Golden oldies: curating SEM specimens. *Collection Forum*, **5**, 17—26.

Golden, J. (1992). Storage for SEM stubs. In *Storage of Natural History Collections: Ideas and Practical Solutions*. *(C.I.* Rose and A.R. de Torres, eds), pp. 271—272. Society for the Preservation of Natural History Collections, Pittsburgh.

Gordh, G. and Hall, J.C. (1979). A critical point drier as a method of mounting insects from alcohol. *Entomological News*, **90**, 57—59.

Hangay, G. and Dingley, M. (1985). *Biological Museum Methods. Vol.2*. Academic Press, Sydney.

Hillyard, P. (1989). Types of preservative. In *British Arachnological Society Member's Handbook*. pp. 1—4. The British Arachnological Society.

Horn, W., Kahle, I., Friese. G. and Gaedike, R. (1990). *Collectiones entomolgicae*. 2 vols. Akademie der Landwirtschaftswissenschaften der Deutschen Demokratischen Republik, Berlin.

Lincoln, R.J. and Sheals, J.G. (1979). *Invertebrate Animals. Collection and Preservation*. British Museum (Natural History)/Cambridge University Press, Cambridge.

Marshall. J. A. and Haes, E.C. (1988). *Grasshoppers and Allied Insects of Great Britain and Ireland*. Harley Books. Essex.

Martin, J.E.H. (1977). Collecting preparing and preserving insects, mites and spiders. *The Insects and Arachnids of Canada*, **1**, 1—182.

Mathias, J. (1990). Housing and maintenance of collections. In *Manual of Natural History Curatorship* (G. Stansfield, J. Mathias and G. Reid, eds), pp. 98—143. HMSO, London.

May, B.M. (1958). Beetle relaxing fluid. *New Zealand Entomologist*, **2**, 29.

Milliron, H.E. (1971). A monograph of the western hemisphere bumblebees (Hymenoptera: Apidae; Bombinae). I. The genera *Bombus* and *Megabombus* subgenus *Botnbias*. *Memoirs of the Entomological Society of Canada*, **82**, 1—80.

Moore, S.J. (1977). Some notes on freeze drying spiders. *Bulletin of the British Arachnological Society*, **4**, 83—88.

Moulds, M.S. (1975). *Embedding Insects and Other Specimens in Clear Plastic*. Australian Entomological Press.

Mound. L.A. and Pitkin, B.R. (1972). Microscopic whole mounts of Thrips (Thysanoptera). *Entomologist's Gazette*, **23**, 121—125.

Noyes, J. S. (1982). Collecting and preserving chalcid wasps (Hymenoptera: Chalcidoidea). *Journal of Natural History*, **16**, 315-334.

Peigler, R.S. (1992). Shipping of pinned insects. *Collection Forum*, **8**, 73—77.

Robinson, G.S. (1976). The preparation of slides of Lepidoptera genitalia with special reference to the Microlepidoptera. *Entomologist's Gazette*, **27**, 127—132.

Sabrosky, C.W. (1966). Mounting insects from alcohol. *Bulletin of the Entomological Society of America*, **12**, 349.

Shear, W.A. and Levi. H.W. (1970). Cleaning museum specimens of spiders and myriapods with ultrasonics. *British Arachnological Society Bulletin*, **6**, 117.

Shelton, S.Y. (1996). The shell game: mollusks shell deterioration in collections and its prevention. *The Festivus*, **28**, 74—80.

Sims, L.L. (1989). How permanent are they? *Insect Collection News*, **2**, *26*.

Taylor, C. (unpublished). A compartmentalised storage system for Mollusca. Hampshire County Council Museums Service.

Tennent, N. and Baird, T. (1985). The deterioration of Molluscs collections: identification of shell efflorescence. *Studies in Conservation*, **30**, 73—85.

Townes, H. (1973). A scientifically designed drawer for insect collections. *Entomological News*, **84**, 53—61.

Upton, M. S. (1991). Methods for collecting, preserving, and studying insects and allied forms, 4th edn. *Australian Entomological Society Miscellaneous Publication*, **3**, 1—86.

Upton, M.S. (1993). Aqueous gum-chloral slide mounting media: an historical review. *Bulletin of Entomological Research*, **83**, 267—274.

Wagstaffe, R. and Fidler, J.H. (1955). *The Preservation of Natural History Specimens. Vol.1: Invertebrates*. Witherby, London.

Walker, A.K. and Crosby, T.K. (1988). The preparation and curation of insects (revised edn.). *DSIR Information Series*, **163**, 1—91.

## Entomological suppliers

**Watkins and Doncaster,** Four Throws, Hawkhurst, Kent TN18, 5ED, UK.

**T.P. Drewitt** (genitalia vials), 15 Palace View Road. London E4 9EN. UK.

**MMarris House Nets.** 54 Richmond Park Avenue. Queen's Park, Bournemouth BH8 9DR, UK.

**Lydie Rigout,** Entomological Equipment, 1 Hillside Avenue, Canterbury Kent CT2 8ET, UK.

**Dr Hildegard Winkler,** Fachgeschaeft für Entomologic, Dittesgasse 11, A—1180 Wien, Austria.

**Andreas Schwabe.** Entomologische Utensilien, Spalenberg 28, CH—4003 Basel. Switzerland.

**Omnes Artes,** Via Castelmorrone, 19—20129 Milano, Italy.

**Bioquip Products.** 17803 La Salle Ave, Gardena, CA 90248, USA.

**Ward's Natural Science Establishment Inc.,** PO Box 92912, Rochester. NY 14692—9012. I·SA.

**Australian Entomological Supplies,** Box 314, Miranda, New South Wales 2228, Australia.



**Plate 21** A sample of shells with 'Byne's disease', a colloquial term for an efflorescence resulting from reactions with organic acid fumes released from wood and other materials used to make storage cabinets (The Natural History Museum).

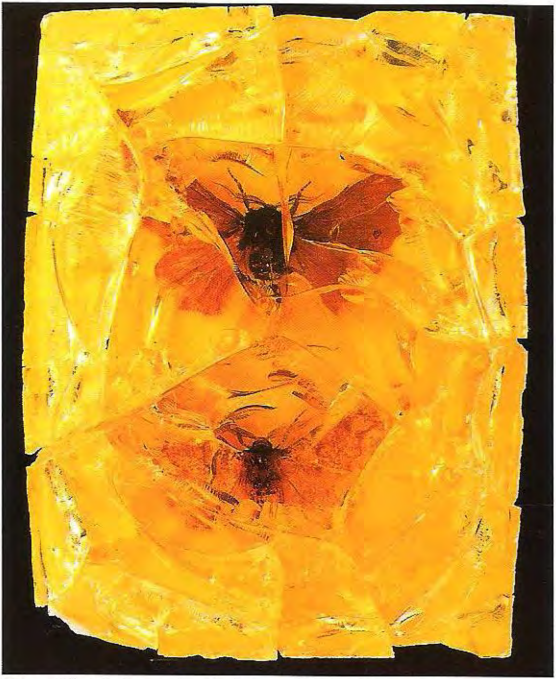


**Plate 22** Moths embedded in a display block of clear plastic resin. The resin has become so cracked and brittle that there is no possibility of recovering either the specimens or the display (The Natural History Museum).




Plate 8 A shell collection with individual specimens enclosed in polythene bags and stored in unit trays (The Natural History Museum).

Plate **9** Rubber stoppers that have perished with age to the point where it is almost impossible to retrieve specimens within the vial without damaging them (The Natural History Museum).

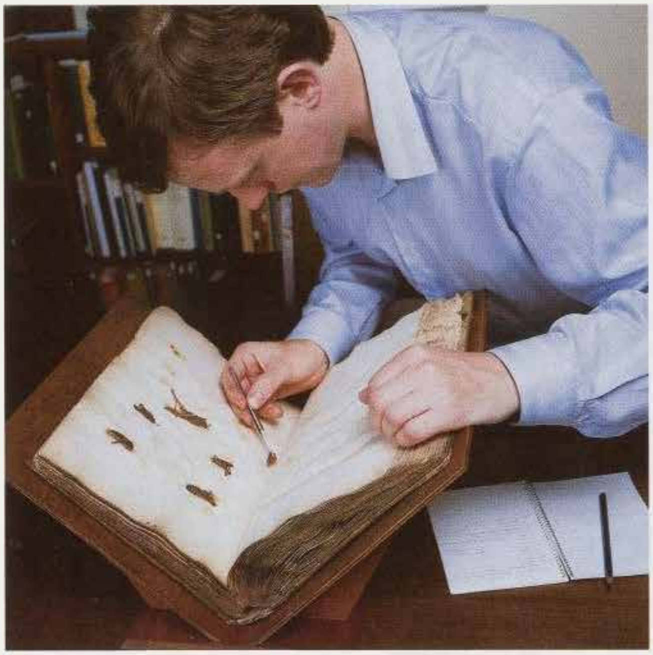


**Plate 6** The Plukenet Collection (c. 1670) illustrates an early technique of pressing insects like flowers, attached to sheets of paper bound as a book. Purchased by Sir Hans Sloane in 1716, this is possibly the oldest insect collection surviving intact. It is housed in the Entomology Department of The Natural History Museum, London and is shown here under remedial conservation (The Natural History Museum).

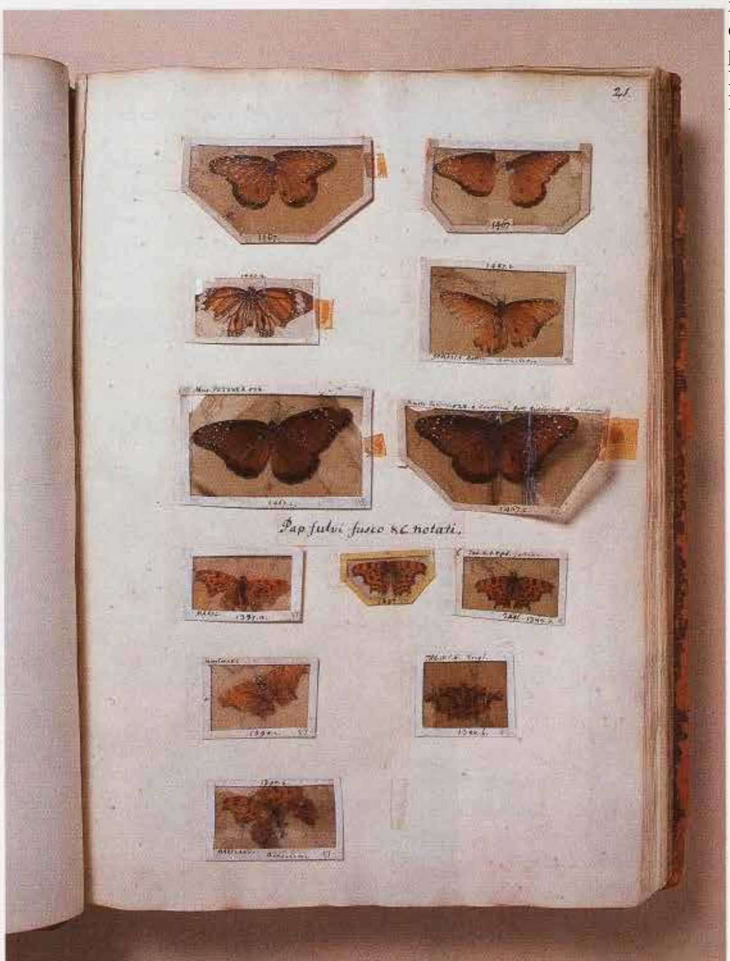



**Plate** 7 Part of the Petiver Collection. This illustrates a slight advance over Plukenet's collection, with the specimens protected by pieces of mica. The collection is housed in The Natural History Museum, London (The Natural History Museum).